



WEST VIRGINIA DEPARTMENT OF ENVIRONMENTAL PROTECTION

West Virginia Save Our Streams Program Level-One Standard Operating Procedures Manual

THE MISSION OF WEST VIRGINIA SAVE OUR STREAM'S IS TO PROMOTE THE PRESERVATION AND RESTORATION OF OUR STATE'S
WATERS BY PROVIDING A BETTER UNDERSTANDING OF THEIR ECOLOGICAL INTEGRITY.

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INTRODUCTION

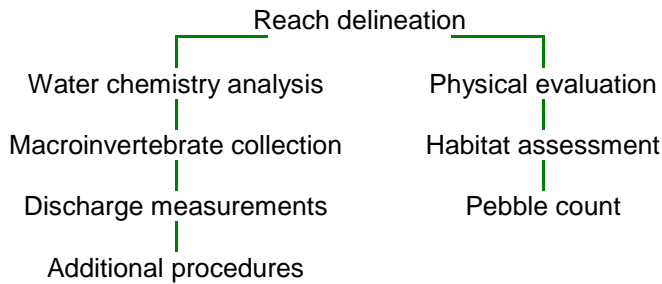
This manual describes the standard operating procedures (SOPs) for completing a **biosurvey** using WV Save Our Streams methods. The SOPs are designed and intended for the assessment of wadeable streams; they consist of three basic elements: (1) water quality analysis; (2) physical characterizations, mostly by observations using the characteristics and conditions described on the survey data sheet; and (3) the bioassessment of the macroinvertebrate community. For more information about general hydrologic and ecological concepts visit the [Volunteer Monitoring Manual](#) section of the West Virginia Save Our Streams' Program website.

Prior to your site visit you should prepare for your survey making sure you have all the necessary equipment. The [equipment](#) should be in working condition and replaced or repaired if needed. Check batteries if electronic equipment such as cameras or GPS units are used. Check the expiration dates of chemicals, make sure all meters are calibrated, all nets are to be secured to their poles and have no tears or holes, and all sample containers should be clean and dry. Review the appropriate topographic maps and/or aerial photos prior to the site visit and possibly have copies on-hand during your survey. Make sure all sites are accessible and that permission to cross private property is acquired prior to your site visit. During surveys, always wear the correct footwear (waders, boots, and closed-toed shoes) and clothes for working in and around the stream. Your footwear should be cleaned prior to visiting the next site to avoid possible transfer of any biological or chemical components. You should have a thorough knowledge of the risks associated with stream survey work in your area and have a corresponding safety plan to reduce those risks. The flow chart below provides the general steps of the stream survey procedure.

CLEAN YOUR EQUIPMENT: Felt glued to the bottom of the wading shoe will improve your traction for walking in the stream; however, felt will also easily transfer certain biological components (i.e. [algae](#), bacteria etc.) if they are not properly [cleaned](#). Equipment and wading apparel should be cleaned between uses and especially if multiple streams are visited.

Before deciding to begin a monitoring or restoration project it is very important for you to describe your [study design](#). Think carefully about the why, what, where, when and how questions, and consider the [quality assurance and quality control \(QAQC\)](#) measures that are necessary to insure accuracy and precision. Your approach should be similar to the [scientific method](#). The questions you ask, the methods you choose, and the way the data is analyzed

and checked should be written into your study design. It's worth taking the time to figure out what you want to do. Your monitoring is much more likely to be successful and sustainable over a longer time, with the right plan.

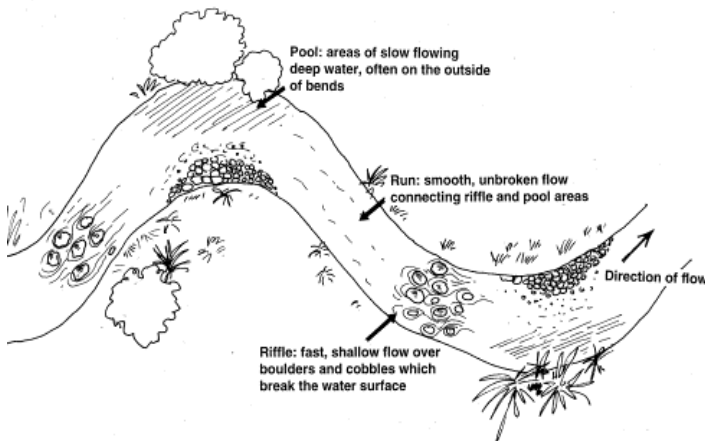


The steps of the procedures can vary slightly, depending upon the number of volunteers available to perform the survey. However most of the physical observations and water quality analysis should occur prior to any sediment disturbance that may be caused by walking in the stream while performing pebble counts or habitat observations, or macroinvertebrate collections.

REACH DELINEATION

The sampling locations on a stream should be a minimum of 50-yards upstream from a road or bridge crossing. Being upstream of these or other types of human encroachments minimizes the effects on stream velocity, channel shape and size, and overall habitat quality. In other words your reach should be as representative as possible of the natural characteristics of the stream. Additionally, no major tributaries (2nd order or higher) should be discharging within the reach.

Once the station is established the length of the reach is determined and set. WV Department of Environmental Protection's (WVDEP), **Watershed Branch** uses a 100-meter reach. Volunteer monitors are encouraged to use the same length but other lengths are acceptable. Most hardware or home stores sell open-reel tape measures of up to 300-feet (100-meter open-reel tape measures are usually available from engineering supply companies). The 300-foot distance is allowable for the maximum reach length. In some cases younger volunteers may be monitoring so it is best to keep them within your line-of-sight. Certain stream-types may meander and have thick vegetation so the entire length of the reach may not be visible. Under these circumstances the length of the reach can be reduced as a safety precaution. The recommended minimum length is 150-feet. The reach should have at least one or more of the following channel features:



RIFFLES have shallow, fast moving water broken on the surface by the presence of coarse substrate such as stacked gravel, cobble and boulders. Its channel shape is variable and often has portions of incline and decline. This channel feature is the best place from which to collect benthic macroinvertebrates.

RUNS are deeper than a riffle with a fast to moderate current and usually no breaks in the surface. The channel shape is relatively consistent with only a slight incline or decline. The substrate is variable but is mostly coarser materials. Your water samples should be collected here, and the width, depth and velocity should be measured within a run. Choose a representative run

that is as close to your x-site as possible. The X-site is the most downstream portion of the reach.

POOLS have deep, slow moving water. The channel shape is generally bowl-like and often some of the bottom substrates consist of finer sediments such as sand and silt. In steep-gradient mountain streams, pools are often deep but may have many areas of very fast velocity and larger substrate. These types of pools are often referred to as steps.

Determine the average **WIDTH AND DEPTH** of the channel features.

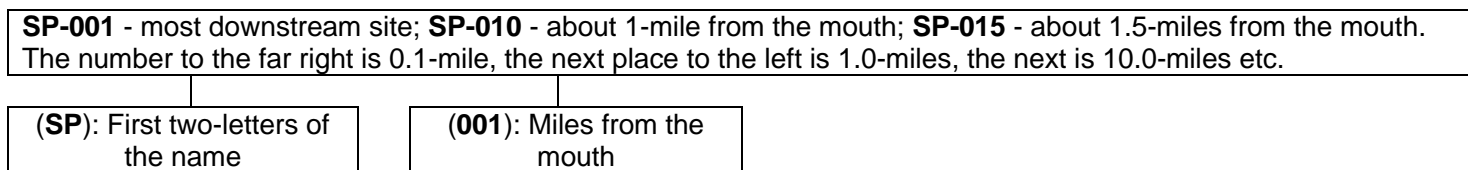
Once the length is determined and set it should remain the same throughout the life of the station. Use flagging or natural features to mark the upper and lower boundaries of the reach. For the first few visits it is a good practice to measure the length of the reach by laying an open-reel tape measure along the banks (not in the stream). The tape measure provides you with many points of reference along the length and is useful for marking the location of

pebble count transects and other notable habitat features such as point bars, islands, or eroding banks that may occur within the reach. The **latitude and longitude (X-SITE)** of your reach is determined at the downstream end of the reach. If you are using a GPS and cannot get a reading at the downstream end, do not move your tape measure once it is in place. Walk up stream until a signal is received and then indicate the location of the signal on your survey data sheet (i.e. middle reach, upper reach etc.). WV Save Our Streams prefers latitude and longitude readings in the degrees-minutes-seconds format.

STATION LOCATIONS

The number and location of your stations depends on what questions your monitoring study has been designed to answer. Table 2 provides some general considerations based upon two types of common monitoring criteria. Prior to selecting the site in the field, use a topographic map to do preliminary selection of sites that meet your criteria. Always visit the site before making final determinations to make sure the site is easily and safely accessible and that it is on public access areas whenever possible.

SITE CODES are important for keeping track of your stations and for reporting purposes. WV Save Our Streams recommends that you use a code consisting of series of numbers and letters that easily designate the site locations and allow for more sites to be added. For example, you have decided on three stations along Spruce Creek. The stations could be coded as follows:



Choose the site that best fit the type of monitoring your group would like to perform. Most volunteer monitoring groups choose sites that determine baseline conditions and will be used to establish long-term or short-term trends. These sites are visited on a regular basis and the information collected is compared to determine if changes are occurring. The other type is for analysis of a particular impact or activity that is occurring on the stream. In this situation the stations are compared against a reference or control to determine the extent of the impact or activity.

Table 1 - Monitoring stations considerations based upon criteria

Characterization study (baseline/trends)	Impact assessment study
(1) Site is typical of the part of the river that interests your group. (2) Site has a variety of characteristics that represents those of the watershed. (3) Site may have some special natural or historical significance. (4) Site may be the location of previous monitoring activity.	(1) A reference or control station is established upstream of the potential impact. In some cases references may be in an adjacent watershed with similar characteristics, or they may be theoretical. (2) The impact station is at, or slightly downstream from the alteration or pollutant. (3) The recovery site is downstream of the impact where it is believed that the stream is beginning to recover from the insult.

How often should you monitor your stations?

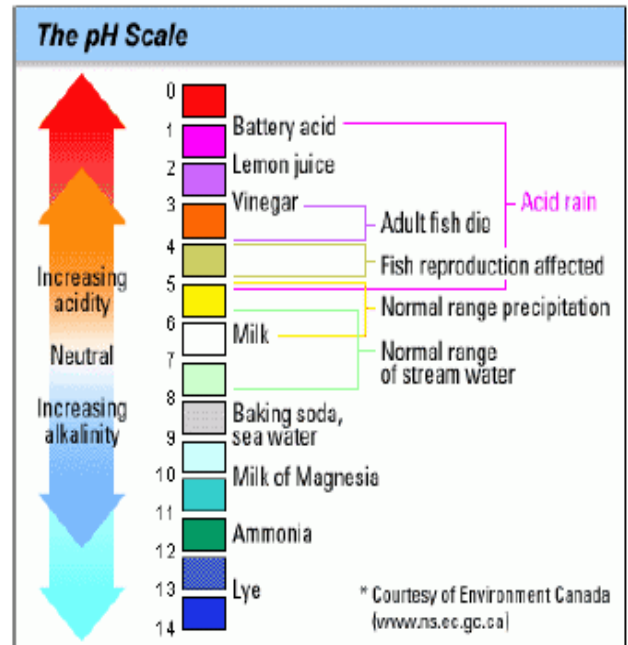
Your study design should help you answer this question. But in general, bioassessment surveys are performed either once or twice within an **INDEX PERIOD**. If there is a reason, volunteers may monitor water quality at a station more frequently such as seasonal or even monthly depending upon the situation. At a minimum the station can be monitored only once within the index period. The WV Save Our Streams index period is from April through October.

WATER CHEMISTRY ANALYSIS

This section describes procedures and considerations for collecting water samples from wadeable stream reaches. For more specific considerations regarding the design of a water quality monitoring program refer to the program's **Volunteer Monitoring Manual** on the WV Save Our Streams website. Water analysis is the first task that should be performed following the delineation of your reach.

Where do I collect my samples?

Water samples should be collected from the most represented portion of the reach, which is usually the run, and as close to the downstream end (**X-SITE**) as possible. If the most downstream end of the reach is a riffle or pool, walk upstream until you encounter a run. Collect the water sample in the deepest section of the run. This may not be the center of the channel depending upon physical features or the curvature of the channel. For example a curved (meander) channel is usually deepest on the outside bend. When wading to your sample location, be careful not to disturb the bottom sediments (or at least keep the disturbance to a minimum). Once you have located the deepest section (thalweg) and have a clean sample container ready, follow the procedure below. In most wadeable streams it is not absolutely necessary to locate the thalweg because the water is well mixed; however, you should never collect your samples from backwater areas.



1. Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also tape your bottle to an extension pole to sample from deeper water if the area cannot be reached.
2. Hold the bottle near its base and plunge it (opening downward) below the water surface. If you are using an extension pole, remove the cap, turn the bottle upside down, and plunge it into the water, facing upstream. Collect a water sample 8 to 12 inches beneath the surface or mid-way between the surface and the bottom if the stream reach is shallow.
3. Turn the bottle underwater into the current and away from you. In slow moving stream reaches, push the bottle underneath the surface and away from you in an upstream direction.
4. Leave about ½ inch air space (Except for DO and BOD samples). Do not fill the bottle completely, so that the sample can be shaken just before analysis. Recap the bottle carefully, remembering not to touch the inside.
5. Fill in the bottle number and/or site number on the appropriate field data sheet. This is important because it tells the lab coordinator which bottle goes with which site.
6. If the samples are to be analyzed in the lab, place them in the cooler for transport to the lab. Some types of samples may need to be preserved to reduce the possible changes that could occur during transport. Your local laboratory can provide you with the necessary preservatives and chain-of-custody forms.

Typically volunteer monitors focus on physical and biological assessments and collect minimal water quality chemistry. WV Save Our Streams recommends that level-two monitors monitor for changes in **pH**, **temperature** and perhaps one or more condition such as **dissolved oxygen** and nutrients (**nitrate/nitrite** and **phosphate**) depending upon the suspected insults. If water quality issues are suspected, chemical monitoring should occur more frequently. Your group should develop a regular schedule to monitor for the pollutant(s) of concern. Below are several water monitoring schedule examples.

1. Seasonal	Once in the winter, spring, summer and fall
2. Monthly	Once from January – December (Winter months are often difficult and dangerous)
3. Water conditions	Based on precipitation events (High, normal, low flows)

WV Save Our Streams does not provide chemical kits, nor can the program pay for laboratory analysis. However, the program has recently purchased several custom **LaMotte kits** and can work with the monitoring group to perform a more thorough chemical analysis at their stations. These kits are only available while working with the WV Save Our Streams Program Coordinator or with one of WVDEP's **Nonpoint Source Program's** Basin Coordinators.

Since there are a limited number of kits, they cannot be loaned or donated to the group unless there is a specific need such as a special study or project. In those cases the volunteer group must show reason for using the kit by providing a written description of the project proposal. The kits are provided at the discretion of the coordinator.

Your water quality monitoring should be based upon the types of land uses common in your watershed. The table below provides several examples of land use activities and the recommended water quality analysis.

Note: This manual does not provide information on the use of any specific chemical kits or meters. The program recommends that whatever type of kit you choose to use, always follow the manufactures instructions and recommendations for its maintenance. If you are using a meter; these must be calibrated at regular intervals and always before using them in the field. When submitting chemical data to WV Save Our Streams always describe the kit you are using, unless the kit was provided by WV Save Our Streams. This description should include the manufacturer, kit-type, range, model number etc.

Table 2 - Water quality monitoring and land use

Land use practices	Recommended water quality analysis
Active construction	DO and BOD, Temperature, TDS and TSS, Turbidity
Forestry harvest	Temperature, TSS, Turbidity
Industrial discharges	Conductivity, pH, Temperature, TDS and TSS, Toxics
Mining	Acidity/Alkalinity, Conductivity, Metals, pH, TDS
Pastureland/Cropland	Bacteria, Nutrients, Temperature, TDS and TSS, Turbidity
Septic systems	Bacteria, Conductivity, DO and BOD, Nutrients, Temperature
Sewage plants	Bacteria, DO and BOD, Conductivity, Nutrients, pH, Temperature, TDS and TSS
Urban run-off	DO and BOD, Conductivity, Nutrients, Temperature

Bacteria – E-coli/Fecal coliform; **BOD** – Biochemical oxygen demand; **DO** – Dissolved oxygen; **TSS** – Total suspended solids; **TDS** – Total dissolved solids; Nutrients (**Nitrates**, **Phosphates** etc.)

Note: Because of the expense and difficulty involved, volunteers generally do not monitor for toxic substances such as heavy metals and organic chemicals such as pesticides, herbicides (agriculture/urban), solvents, and PCBs (industrial/urban). They might, however, collect water samples for analysis at accredited laboratories.

FILTERING WATER SAMPLES

Filtrating involves forcing water through a membrane filled with tiny holes, about 25 microns across (¼ of a human hair). The filter removes suspended solids from the water, and in fact, the weight of the solids caught in the filter is one common analysis parameter: total suspended solids (TSS). Such solids can be virtually invisible to the naked eye, but not dissolved, so they'll eventually settle out if the sample is left undisturbed.

Polluted coalmine water can have tiny bits of iron hydroxide (a.k.a. **YELLOW BOY**) suspended in it. In a water sample fixed with acid but not filtered in the field, this iron hydroxide will dissolve into the water before it arrives at the laboratory, which will give incorrect test results for metals. Alternatively, in a sample that is not acidified, dissolved oxygen can cause dissolved ferrous iron to oxygenate into ferric iron en route to the lab. Unless the insoluble ferric iron is filtered out at sampling time, the lab can't determine how much ferrous vs. ferric iron the water actually contained when it was sampled. Ferrous iron oxygenation also consumes acid, so certain laboratory acidity or pH measurements will be wrong.

Although filtering water samples eliminates these errors to some extent, it requires special equipment and training. Many volunteer groups leave filtering to the analysis laboratory, accepting any inaccuracies that creep in beforehand. Ultimately, the decision to filter relies on a group's capacity and requirements, and it should be discussed with the laboratory that receives the water samples. The recommended preservative for various constituents related to water monitoring are based on a wide variety of references and information.

PHYSICAL EVALUATION

This portion of the survey includes a wide variety of observations using all senses, making several judgments based on established rating descriptions, as well as some collection and measurement procedures. In this section we discuss general physical observations. These observations should be completed prior to or immediately following the water quality analysis.

Table 3 provides a list of the conditions that are assessed as wells as some general guidelines regarding what certain characteristics indicate. Water observations are made in a run or riffles, sediment observations are also made in a run or riffle and **benthic algae** observations are made in a riffle. These observations should be made multiple times throughout the reach to make sure conditions are consistent. You should write comments on the survey data sheet if any notable differences within the reach are observed. Note: The University of Maine's **field guide to aquatic phenomenon** provides a good overview (with pictures) of many conditions described in Table 3.

Table 3 - Stream physical condition characteristics

Water Colors

Brown: Usually caused by sediment in the water. Some muddiness (brown color) is natural after storms, but if the condition persists look for an activity upstream that has disturbed the soil such as construction sites, logging, storm water runoff from roads or urban areas, or agricultural activities such as cattle in the stream.

Black: Usually caused by coalmine drainage, tar or sometimes waste material from road construction.

Green: Usually due to an algae bloom caused by excessive nutrients in the water. The source could be sewage, fertilizers from farms, homes or golf courses or waste from animal feedlots.

Multi-colored sheen: Can occur naturally in stagnant waters, but a sheen that is moving or does not break up easily may be an indication of oil pollution. The source could be runoff from streets or parking areas or illegal dumping. In some areas the use of all-terrain (ATV's) vehicles may contribute to stream oil pollution.

Orange or red: Usually associated with mine drainage (high iron content in the stream).

Tea colored: Usually associated with wetlands.

White or gray: Can be caused by runoff from landfills, dumps or sewage.

Sediment Colors

Brown: An indication of silt deposits from sediment sources. Most stream bottoms are normally brown in color.

Black: This deposit can occur naturally in heavy organic soils but can also be due to fine coal particles, tars, ashes, sludge etc.

Green: Possible indication of excessive algae growth from organic (nutrient) enrichment sources.

Orange, yellow or red: A coating of flocculates on the sediments is usually due to polluted coalmine drainage.

White or gray: A white cottony mass is a sewage fungus common to organic polluted waters. An even coating of white or gray flocculates may be metals (aluminum) precipitated out of solution from contamination due to mine drainage.

Water/Sediment Odors

Collect your sediment and water sample separately to assess the odors.

Rotten eggs: This strong sulfur-like odor can be an indication of sewage pollution or polluted coalmine drainage.

Musky: This slight organic odor is often natural, but in some cases may indicate nutrient enrichment from organic waste products or sewage contamination.

Oily: This odor may indicate pollution from oil and gas wells.

Chemical: There are a wide variety of chemical odors usually the result of industrial discharges, solvents and detergents.

Algae Color

Algae color varies from brown to dark green in most streams and rivers; although color is a noticeable condition of the algae it is not a particular indicator of the types or of the condition represented by the algal community.

Algae Abundance

Coverage in a riffle is estimated based upon the following: none, scattered, moderate or heavy. A heavy coating of matted and floating algae is often an indication of nutrient rich conditions caused by excess nitrogen and phosphorous.

Algae Growth habit

The growth habit characteristics are critical to understating the algae. Most stream algae will be evenly coated on the rocks and have a smooth or slimy texture; other types will be filamentous and have a hairy texture; and others will be matted. Matted algae are easily removed from the surfaces by slowly scraping with your fingers. If the algal community is mostly matted pieces will come off in junks like carpet when it has been removed from flooring.

Foam occurs naturally due to the decomposition of leaves (this foam is generally less than three inches high and cream colored). Excessive white foam may be due to detergent pollution.

HABITAT ASSESSMENT



The **habitat assessment** process involves rating and scoring many different **habitat conditions** as optimal, suboptimal, marginal or poor based upon criteria (descriptions and a rating scale) included on the survey data sheets. The optimal category is a description of conditions that meet natural expectations; suboptimal includes descriptions of criteria that are less than desirable, but satisfies expectations under most circumstances; marginal is a description of moderate levels of degradation with severity at frequent intervals throughout the reach; and poor are descriptions of criteria for streams that have been substantially altered with severe degradation characteristics.

Descriptions of the habitat conditions are provided below. For additional information refer to Chapter 5 of the US EPA's **Rapid Bioassessment Protocols** Manual. The conditions here should be applied when describing the habitats of rocky-bottom streams and rivers.

EMBEDDEDNESS refers to the extent to which rocks (gravel, cobble, and boulders) are surrounded by, covered, or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become

embedded, fewer living spaces are available to macroinvertebrates and fish for shelter, spawning and egg incubation. To estimate the percent of embeddedness, observe the amount of silt and sand sediments overlying and surrounding the larger gavel and cobble size particles. You should base your embeddedness assessment on the composition of the materials that you observe. Embeddedness is always evaluated in the riffles used for your macro-invertebrate collections. In most cases the best person(s) to comment about this condition is the person(s) collecting macroinvertebrates. If cobble and gravel are easy to remove from the riffle and there is little sand or silt either in the net or suspended during collections, embeddedness is minimal. In some cases chemicals can cement (armoring) the substrate together and cause severe embeddedness.

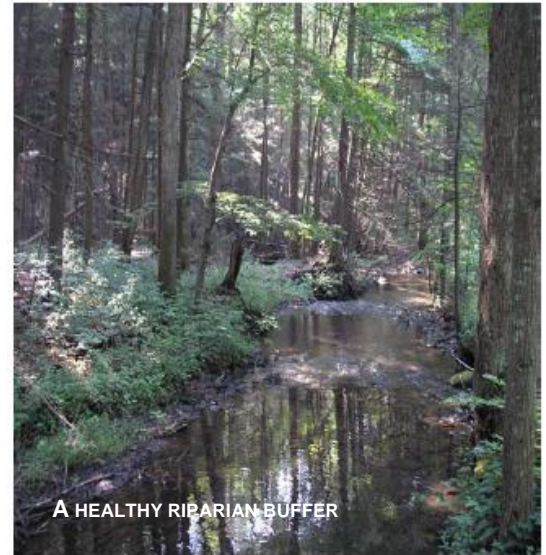
SEDIMENT DEPOSITION is an estimate of the amount of sediment that has accumulated and the changes that have occurred to the stream channel as a result of deposition. Deposition occurs from large-scale movement of sediment. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends. High levels of sediment deposition are symptoms of an unstable and continually changing environment that becomes unsuitable for many organisms. Sediment deposition should be rated throughout your reach and should not be confused with embeddedness. Sediment deposition is probably the most difficult condition to assess. It is a natural process and bars often form in streams that are very stable and have little sediment from the surrounding land or few problems with erosion. When assessing this condition look for indicators that are unusual or beyond what is expected to be normal for the stream. The most effective way to learn is to view many different stream types representing both degraded and natural conditions. In most cases island formation, especially in small streams (1st through 3rd order), is an indication of excessive deposition. The most common cause for unusual or un-natural deposition in most streams is human encroachment (i.e. structures such as bridges, roads, culverts etc. too close to the stream or built so that the stream is narrowed) and bank erosion. Steep sloping banks with exposed surfaces are more likely to erode. Undercut banks can often erode but are sometimes very stable if covered with vegetation, tree roots and rocks. Look for deposition around eroding banks, especially if they show bare soils consisting mostly of fine materials (fine gravel, sand and silt). Hard surfaces no matter how steep or undercut are less likely to erode.

The remaining conditions are assessed on the left and right sides. Note: The **LEFT** and **RIGHT** sides are determined by looking downstream.

The **BANK STABILITY** condition evaluates whether the stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks, and are therefore considered to be unstable. Signs of erosion include crumbling, un-vegetated banks, exposed tree roots, and

exposed soil. Eroded banks indicate a problem of sediment movement and deposition, and suggest a scarcity of cover and organic input to streams.

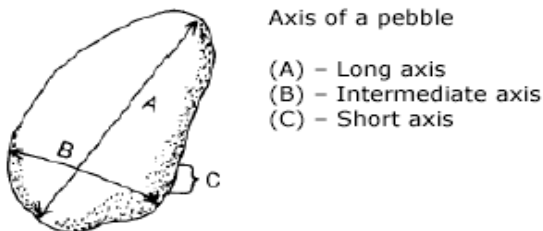
RIPARIAN BUFFER WIDTH is an estimate of the width of natural vegetation from the edge of the stream bank out through the riparian zone. A relatively undisturbed riparian zone supports a robust stream system; narrow riparian zones occur when roads, parking lots, fields, lawns, bare soil, rocks, or buildings are near the stream bank. Residential developments, urban centers, golf courses, and rangeland are the common causes of anthropogenic degradation of the riparian zone. Conversely, the presence of old fields, paths, and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to altering the riparian zone and may be given relatively high scores. Riparian buffers are the most valuable protection a stream system has against outside influences. In most cases healthy riparian directly reflects upon the condition of the stream unless the source of the insult is a specific pollutant. Enhancement of the riparian buffer by re-planting native grasses, forbs, shrubs and trees is the first step in the recovery of the stream back to a more natural condition. Below is a list of just a few of the many benefits a healthy riparian buffer provides.



1. Provides organic material as food for macroinvertebrate, fish and wildlife.
2. Supplies large and small pieces of woody debris that provide habitat for fish, macroinvertebrates and amphibians.
3. Alters how sunlight reaches the stream and is an important temperature moderator.
4. Stabilizes stream banks and reduces erosion.
5. Filters sediment and materials from overland runoff and roots of many plants traps and holds the sediments.
6. Absorbs nutrients from overland and sub-surface flows.
7. Reduces the impacts of flooding through temporary storage, interception/diversion and slow releases (especially wetlands) from heavy rains.

PEBBLE COUNT

The composition of the streambed and banks is an important facet of stream character, influencing channel form and hydraulics, erosion rates, sediment supply, and other parameters. Observations tell us that steep mountain streams with beds of boulders and cobbles act differently from low-gradient streams with beds of sand or silt. You can document this difference by collecting representative samples of the bed materials using a procedure called a **Wolman pebble count**. This method requires a person with a metric ruler who walks through the stream and a note taker who remains on the bank with the field book or survey data sheet. The note taker records the count is recorded by size classes or categories similar to those shown in Table 4. Pebble counts can be made using grids, transects, or a random step-toe procedure.



Select the portion of the reach that you wish to measure (this may be the entire reach or riffles only). For stream characterization, sample pools, runs and riffles in the same proportions as they occur in the study reach. For other purposes, it may be more appropriate to use a more random method. The professional SOP recommends a minimum of 100 particles to obtain a valid count. WV Save Our Streams ask that volunteers using this procedure collect a minimum of 50 particles.

Table 4 - Pebble count size classes

Categories	Description (size)	Size range (mm)	Categories	Description (size)	Size range (mm)
Silt/clay	Slick/smooth	Determined by feel	Cobble	Tennis ball - basketball	65 - 256
Sand	Very small grainy	< 2	Boulder	Basketball – car size	> 257
Fine gravel	Pea – golf ball	2 - 24	Bedrock	Large solid surface	NA
Coarse gravel	Golf ball – tennis ball	25 - 64	Woody debris	Sticks, leaves etc.	NA

Collection procedure: Start at a randomly selected point near the downstream end of the reach. Start on the shoreline and take steps across the stream. Averting your gaze, pick up the first particle touched by the tip of your index finger at the toe of your wader. Measure the intermediate axis (neither the longest nor shortest of the three mutually perpendicular sides of each particle picked up). Measure embedded particles or those too large to be moved in place. For these, measure the smaller of the two exposed axes. Call out the measurement. The note taker tallies it by size class, repeats it back for confirmation and also keeps count. Continue across the channel slightly upstream in the direction of the opposite bank and repeat the process, continuing to pick up particles until you have the requisite number (100 or more) of measurements. Traverse across the stream perpendicular to the flow or in a zigzag pattern.

MACROINVERTEBRATE COLLECTION AND ASSESSMENT

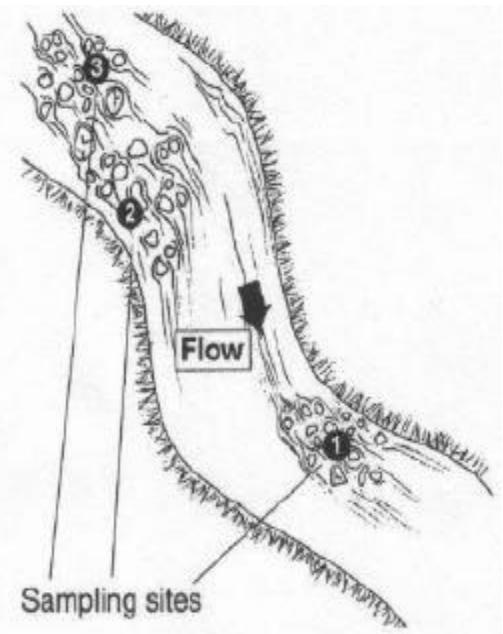
The groups of aquatic animals found in leaf packs, rocks, woody debris and other areas of streams, rivers, ponds and wetlands are collectively called **BENTHIC MACROINVERTEBRATES**. Benthic refers to the ability to cling to bottom surfaces such as rocks, leaves or roots. Macroinvertebrates are animals without a backbone that can be seen with the naked eye. These bottom-dwelling animals include crustaceans, mollusks and annelids but in many aquatic environments, most are larvae of aquatic insects. Macroinvertebrates are an important link in the food web between the producers (leaves, algae) and higher consumers such as fish. They are the key indicators of biological integrity in a wide variety of aquatic environments.

Depending upon the stream type a variety of methods and equipment are used to collect benthic macro-invertebrates from wadeable streams. In rocky-bottom streams, WV Save Our Streams recommends using a two-pole screen-barrier net (or two-pole kick-net) or a single pole **rectangular style kick-net**. Groups may purchase other nets as needed but only the two-types discussed here are recommended for rocky-bottom collections. Ultimately, the nets chosen depend upon the goals and objectives of the volunteer monitoring program.

COLLECTING BENTHIC MACROINVERTEBRATES

Benthic macroinvertebrates live in a wide variety of aquatic environments. In lakes, wetlands and large river systems they are common in shallow edge micro-habitats along shorelines in tangles of vegetation, roots, and leaves, in gravel shoals or along rocky and undercut banks; some kinds burrow in mud and sand of shallow flowing water. In streams and rivers with swift-flowing water they are more common and diverse in rocky areas (riffles), but are also found in runs, which are sampled when riffles are not present. The collection procedure described here is designed for rock-bottom streams from riffle habitats. For information about the muddy-bottom bioassessment procedure visit WV Save Our Streams Volunteer Monitoring Manual web page. The procedure is described in Chapter 4: Macroinvertebrate and Habitat beginning on page seven. The number of samples collected depends upon the type and size of the net. Standard two-pole kick-nets are about 3-feet wide (1-meter²) so usually, three-samples are adequate. These types of nets can only be used in riffles and runs. Other nets such as the rectangular kick-net or **D-nets** are also used. These nets are much smaller but more versatile than the two-pole net. However, more samples are necessary in order to collect an adequate representation of benthic macroinvertebrates. For example if you use the rectangular kick-net, six to eight samples should be collected.

1. **CHOOSE THE BEST HABITATS:** Your goal is to collect macroinvertebrates from three different riffle areas. (If the riffles are as wide as the stream then multiple samples could be collected within the same riffle.) The riffles should have different characteristics (i.e. different composition but mostly cobble and gravel and different velocities). Often different types of riffles hold different varieties of macroinvertebrates, so to properly assess the biological conditions you need to collect a representative sample. Of course your choices of habitats will ultimately depend upon what your reach provides. Once you've chosen your location always approach from the downstream end, sampling the site farthest downstream first. This approach insures that the sample is representative of its location and reduces the chances of biasing your second and third sample.



2. **GET INTO POSITION:** Select an area approximately the same width (or slightly less) than the width of your kick-net. Most two-pole screen-barrier kick-nets are about 3-feet to 1-meter wide. The width of the single-pole rectangular kick-net is about $\frac{1}{4}$ to $\frac{1}{2}$ meter. If you are using a smaller diameter-net you will need to collect more samples (About two-three samples using the rectangular kick-net is equivalent to one sample with the screen-barrier kick-net). The net holder will place the net snugly against the bottom of the streambed; rocks can be removed if necessary to make sure you have a close-fit. Once you are satisfied with the position, line the front of the net with rocks heavy enough to hold the net in-place. However, be careful to choose rocks that are not too heavy, or too wide or high. Large rocks will damage the net and will influence how the macro-invertebrates flow into the net thus making capture less successful. The net holder now tips the kick-net backward at about a 45-degree angle from the water's surface. This provides greater surface area and more even flow into the net. If the net is held too high some of the macroinvertebrates will wash around the sides and not be captured in the net. While holding the kick-net backwards the net-holder must make sure that water does not wash over the top of the net. Have one 3 $\frac{1}{2}$ to 5-gallon bucket ready before you begin collecting the sample. The bucket will be needed during sample collection.

3. **BEGIN DISTURBING THE STREAMBED:** The second person approaches the sample area from upstream and determines the approximate sample-size. Once the area is delineated, the sampler begins disturbing the streambed directly in front of the net. The process starts with rock rubbing. First, pick-up all large rocks (cobble size and larger) and inspect them. You are looking for snails, clams and caddisfly cases. These animals often cling very tightly to the rocks and are not removed by just a simple rub with the hands or a small brush. If the rocks have any of these animals, remove them from the rocks and place them inside the bucket. Move the rocks you picked-up towards front of and slightly into the net; brush all sides of the rocks with your hands or a small vegetable-brush to dislodge other clinging macroinvertebrates. Continue the rock rubbing process until the larger sized stones have been thoroughly cleaned. If the rocks cannot be lifted from the streambed, simply rub them where they lay. As much as possible the rock rubbing should proceed from the upstream portion of the sample area towards the front of the net. After the rocks are rubbed they should be placed aside (outside of the sample area) so they are not rubbed a second time. Some volunteer programs choose a timed approach to rock rubbing since this is a rather intensive and somewhat time consuming step. The recommended time frame for adequate rock rubbing is two-four minutes depending upon the abundance of cobbles and boulders within your sample area. If you choose a timed approach you should make a note on the survey data sheet and record the time frame you use.



After you are satisfied that all or most of the larger rocks have been cleaned you will disturb the remainder of the streambed using a kicking method. Position yourself upstream from the net inside your sample area and begin shuffling your feet back and forth from one side of the sample area to the other. Slowly move towards the net while kicking from side to side. The action dislodges macroinvertebrates from smaller size gravels and also disturbs those that might be buried themselves down into the soft bottom sediments. While you're moving from side to side and forward you are also pressing your feet downward in an attempt to make a depression in front of the kick-net. If you have chosen a timed approach then you should limit your kick-time as well. The recommended kick-time is two minutes, so your entire sample should take about six minutes to collect.

4. **REMOVE THE KICK-NET FROM THE STREAMBED AND CAPTURE THE COLLECTION:** This is a very important step; since the sample collection is laborious you do not want to lose any of the macroinvertebrates collected by sloppy procedures here. Very slowly remove the rocks that have acted as anchors to hold the kick-net in place, rub them off while you remove the rocks, or you may choose to rub them before using them to anchor the kick-net in place. While the net-holder grabs and holds the top of the net in-place the kicker grabs the bottom edge of the net near the handles. The net is removed with a scooping motion, the kicker moved slightly forward and

upward while the holder keeps the net steady so that no macroinvertebrates are lost from washing over the top of the kick-net. Both persons then pick-up the net and roll it into a loose cylinder, securing the ends and taking it to the shoreline. The bucket that was used earlier in the procedure should be at the ready to accept the contents of the kick-net.

5. **PLACE THE NET INTO THE BUCKET:** Slightly unwind the net so that it fits inside the bucket. With a smaller bucket or a spray bottle, wash the contents of the kick-net into the bucket. It will take several minutes and several washes to knock loose most of the macroinvertebrates. Between each attempt, remove the net and check for macroinvertebrates that have not been dislodged. Often these hardy clingers are found near the edges of the kick-net along the bottom-side and in the seams of the net. Be sure to check the opposite side for macroinvertebrates that may have crawled in an attempt to escape. You must be very careful not to overfill the bucket. If the bucket begins to fill with stream water more than about two-thirds its heights, remove some of the water by seining it through the kick-net (hold the net tightly on the bucket and pour off the water) so that the water is poured off and the macroinvertebrates remain in the bucket. The process is complete when you are satisfied that the kick-net has been thoroughly washed and most of the macroinvertebrates are now in the bucket.



6. **REMOVE THE CAPTURED MACROINVERTEBRATES FROM THE BUCKET AND BEGIN SORTING:** The goal of this step is to remove all captured macroinvertebrates so that they can be observed, identified and counted. WV Save Our Streams recommends that you use several shallow white trays. The best way to start is by trapping the macroinvertebrates as they are poured from the bucket. Before starting the steps below, remove all larger materials that may have been collected with your sample from the bucket. Make sure to check these for macroinvertebrates before they are discarded. At certain times of the year leaves and other debris are very plentiful in the stream and this material must be sorted. (It is common to find many kinds of macroinvertebrates in leaf-packs; this material is one of their favorite places to live.) The best way to deal with the leaves is to remove as many as possible, place them in smaller bucket or container and wash them to remove the macroinvertebrates.

Pay close attention to the leaves that appear chewed and have begun to decay. Newly fallen leaves are less likely to have many benthic macroinvertebrates.

Removing (seining) can be completed by using a second bucket with your kick-net on top, and then pouring the captured organisms over the net so that they are trapped. The pouring is stopped periodically so that the macroinvertebrates can be removed from the net and placed into the collection trays. Small forceps are the best tool for this job; however the macroinvertebrates can also be removed by hand. Another method is to use a **sieve bucket** (wash bucket) or **EZ-STRAINER**. The EZ-strainer is highly recommended, and is available in limited-quantity from the WV Save Our Streams Program. The EZ-strainer comes in a variety of mesh sizes (some even finer than 500-microns) and fits nicely inside a 3 ½ or 5-gallon bucket. The bucket used to capture your collections can easily be poured into the second bucket and the macroinvertebrates are removed from the EZ-strainer.

The collection procedures are repeated three or more times, one for each sample collected. Volunteers may choose to sort each sample separately or combine all three samples and sort them at one-time. The latter is highly recommended since the bioassessment procedure is based on a composite sample. In certain (rare) circumstances samples may need to be counted, sorted and identified separately to compare different portions of the reach. These comparisons are done only when there is a suspected insult to the macroinvertebrates such as a habitat alteration or a suspected chemical that may not be affecting the entire reach. Collection, sorting, counting and identification are the most intensive tasks of the survey procedures.

Note: If your organization decides to complete independent surveys that include **aquatic collections**, you must apply for and receive a **SCIENTIFIC COLLECTION PERMIT** from the WVDNR. [**\$20-2-50**]

MACROINVERTEBRATE COUNTING, IDENTIFYING AND SORTING

Sorting macroinvertebrates from survey samples (a procedure often referred to as "bug picking") is an extremely important step in the biological research. The quality of the work performed by the "picker" influences the quality of subsequent processes, such as identification and data analysis. A competent "picker" must be able to recognize the morphological diversity of aquatic organisms, as well as the various methods these organisms may use to hide themselves from predators. The outcome of the final study may be affected, even if only a few organisms are overlooked during the picking process. Table 5 provides a list of materials and supplies needed for this work.

Table 5 - Recommended supplies for bug picking and identification

1. Pans (larval trays): contains sample during the sorting process.
2. Gridded tray: a white enamel or plastic tray used to separate family groups. The tray can be marked into equal size grids ranging in size from 1 x 1 to 3 x 3 inches. The grids help to estimate the numbers. Ice-cube trays and **plastic craft organizers** are also effective tools.
3. Forceps: fine tipped forceps are used to remove the organisms from the debris and to manipulating specimens during examinations to determine identification.
4. Magnifier: an optical aid to illuminate and magnify the sample during the picking process.
5. Various taxonomic keys (e.g. **dichotomous keys**, picture or **illustrated keys**, and other guides), which provide some family-level descriptions.

Once the collections are placed into larval pans you will need to sort and identify. At times this can be very difficult in the field so your group may want to consider using alternate procedures such as preservation and sub-sampling. This is a very important part of the survey procedure and it requires a great deal of time and some expertise. The procedure can vary based upon the contents of your collection. **USE THE STEPS BELOW AS A GUIDE.**

1. Add a couple of inches of water to your main sorting tray and the others you will be using. Spread the sample out over the bottom of a white tray. Spend a little time watching the macroinvertebrates. See how they move and look at the different shapes and colors (the colors change when they are preserved).
2. Pick through your sample in the sorting tray. Use a pipette, tweezers, spoon or brush to transfer your macro-invertebrates to the wells in the ice cube tray, craft organizer or smaller tray. Place animals belonging to the same group in the same portions of the tray. In some cases it may be simpler to sort through one order at a time, especially if multiple kinds are probable. For example, **EPHEMEROPTERA** (Mayflies), **PLECOPTERA** (Stoneflies) and **TRICHOPTERA** (Caddisflies) can be very abundant in healthy streams and these orders as well as **DIPTERA** (True flies) are likely to have multiple families present.
3. For the first 10–20 minutes, transfer any animal that you see from the sorting tray into the other trays. For the last 10–20 minutes, look particularly for animals that are uncommon. Fast moving macro-invertebrates will be obvious but some will only start to move after 10 minutes or so. If after 30 minutes you find an invertebrate you haven't seen before, sort for another 10 minutes until you find no new families.
4. There are many key guides available to identify your macroinvertebrates. A 10x magnifying loupe, magni-cube or low power binocular microscope is useful for looking closely at the animals.
5. Estimate the abundance or count the number of each kind of animal in the tray sections. As you look for families and count or estimate abundance keep track of your tallies on the survey data sheet. Use a pencil so you can erase or scratch through your numbers when different families are encountered or your abundance changes. If you find a macroinvertebrate you cannot identify, record this on your result sheet, giving a brief description of what you found. When you finish, return your collections to the stream as close to the collection sites as possible.

CALCULATING YOUR STREAM SCORE

After the sorting and identifications is complete, the macroinvertebrate are assessed using three **METRICS**, which are converted into an overall stream score using a simple point system. First, transform your abundance rating into numbers: (**A** = 6; **C** = 3; **R** = 1) then follow the instructions to complete all the necessary calculations.

Level-one metric calculation example

Insect groups	Abundance	Tolerance Value	Tolerance Score	Taxa
Stoneflies	1	2	2	1
Mayflies	3	3	9	3
Case-building caddisflies		3		
Net-spinning caddisflies	1	4	4	2
Common netspinner	6	5	30	1
Free-living caddisfly		3		
Dragonflies				
Damselflies		7		
Riffle beetle	3	4	12	1
Water penny	3	3	9	1
Other beetles		6		
True bugs		8		
Hellgrammite (Fishfly)	1	3	3	1
Alderfly		6		
Non-biting midge	3	9	27	1
Black fly	3	6	18	1
Crane fly		4		
Watersnipe fly	1	3	3	1
Other true flies		7		
Non-insect groups	Abundance	Tolerance Value	Tolerance Score	Taxa
Water mite		6		
Crayfish	3	5	15	1
Sideswimmer (Scud)		5		
Aquatic sowbug		7		
Operculate snails	1	4	4	1
Non-operculate snails	1	7	7	1
Clams		6		
Mussel		4		
Aquatic worm	1	10	10	1
Leech		10		
Flatworm		7		
Other (describe)				
Totals	Total Abundance		Total Tolerance	Total Taxa
	31		154	17

See the example calculation (left). For additional information refer to the survey data sheet in Appendix C.

- Biotic Index** - Multiply the abundance number by the tolerance to calculate the tolerance score. Add the entire tolerance score column and the relative abundance column. Divide the total tolerance by the abundance total.
- Total Taxa** - Calculate the total number of kinds (taxa).
- EPT Taxa** - Calculate the total number of kinds (taxa) from the stoneflies, mayflies, and caddisflies.

WV Save Our Streams provides on-line [spreadsheets](#) for calculating these metrics. If a group prefers not to calculate the stream score, the coordinator will perform the calculations when the data is reviewed.

Tolerance values (TVs): TVs are general numbers that represent the tolerance of most of the group. It should be noted that tolerance ranges vary for certain Mayflies (a few kinds range from 4-5), Beetles (a few kinds are 5), other True flies (a few kinds are 6; Net-wing midge TV = 2) and Operculate snails (a few kinds are less than 5).

Metrics	Results	Points	10	8	6	4	2
Total Taxa	17	8	> 18	18 - 15	14 - 11	10 - 7	< 7
EPT Taxa	7	6	> 10	10 - 8	7 - 5	4 - 2	< 2
Biotic Index	4.96	6	< 3.5	3.5 - 4.3	4.4 - 5.6	5.7 - 6.5	> 6.5
Stream score		20	Integrity Rating				
			Optimal	Suboptimal	Marginal	Poor	
			> 24	24 - 19	18 - 13	< 13	

MEASURING VELOCITY

The stream's velocity (commonly called flow) is modified by conditions along and around the stream, such as:

- Structures, such as dams and weirs, in the waterway;
- Removal (diversion) of water for use in irrigation, industry and households;
- Rainfall, snow melt, and water releases from dams, power stations and industry;
- Entry of groundwater;
- Evaporation ([evapotranspiration](#)) and leakiness of the riverbed and banks.



The size of a waterway and its flow rate affect its water quality. For example, discharges containing contaminants will have less effect on large swiftly flowing rivers than on small slow streams. This is one reason for measuring flow - to work out the load of contaminants and sediment the waterway is carrying. Because discharge can have a significant effect on water quality, it is important that it is recorded at the time of sampling and, if possible, during the previous few days. It is particularly valuable to know if flows are at low, moderate or high level and if the level is rising or falling. This is because the concentrations of nutrients, turbidity and contaminants tend to be higher when the stream level is rising than when it is falling. There are three ways to measure discharge. A simple method is to see how fast a floating object travels downstream over a chosen distance. This is called the float method. Secondly, flow data can be obtained from [US Geological Service](#) or the [State Emergency Management Office](#), if your site is near a gauging station. Thirdly, the velocity head rod (VHR) method can be used.

The **FLOAT METHOD** is easy to understand and something most of us have done as children. You simply float an object on the water and measure the time it takes to travel a set distance. The equipment you will need for this method includes:

- Foam golf ball (preferred), tennis ball, apple or orange peel
- Net to catch the ball
- Open-reel tape or survey rod
- Stopwatch

Procedure: Mark off a 20-foot length of the stream. Choose a section that is relatively straight and free of vegetation or obstacles. Avoid areas with a culvert or bridge because those structures will modify the true flow. If the flow is very slow, mark out a shorter distance. Position a person at each end of the section. Place the ball on the surface near the middle of the stream at least two-feet or more upstream of the end of the tape so it has time to come up to water speed. When the ball is in line with the beginning of the tape, start the stopwatch. Stop the watch when the ball gets to the end of the section. Repeat the procedure at least three times at this site and average the results. To calculate the water velocity, divide the distance travelled in feet by the time taken in seconds. Then multiply by a correction factor to compensate for the variability in velocity with depth and across the channel, i.e. water will move slower at the edges than in the middle and at different speeds within the water-column.

See the example below for a rocky-bottom stream.

Distance travelled = 20 feet	Velocity = $20 \div 18 = 1.1$ ft/sec	Depth = 0.6 ft; width = 10 ft
Average time = 18 seconds		Cross sectional area = 6.0 ft ²

Discharge = $1.1 \times 0.8 \times 6.0 = 5.3$ ft³/sec (cfs)

Note: The US EPA's Volunteer Manual recommends using a correction factor in the calculation based on stream type. The factor for rocky-bottom streams is **0.8** and the factor for sandy/muddy bottom streams is **0.9**.

The **VELOCITY HEAD ROD (VHR)** is a fast and inexpensive method of measuring velocity in a stream. The rod can be a yardstick or meter stick, or it can be made using a 3 - 6 foot long, very thin, piece of wood. A 26-gauge copper sheet is sometimes fastened to the cutting edge to protect it from abuse. Mark a scale in ½ inch increments on the rod, starting with zero at the bottom of the rod and stopping at 18 inches. Steps to find the flow rate with a velocity head rod are as follows:

- Place rod in the water with sharp edge upstream. Measure stream depth on scale.
- Place rod sideways in the water. This will create turbulence and the water will "jump" or rise above its normal depth; velocity is proportional to this jump.

- Measure depth of turbulent water next to rod. Subtract stream depth from the turbulent depth reading to obtain the "jump height," or velocity head in inches.
- Find the stream velocity in feet per second from the table provided on the survey data sheet.
- Determine the stream velocity at intervals across the stream and average them to obtain the average stream velocity in feet per second.

CROSS SECTIONAL SHAPE varies with position in the stream, and discharge. The deepest part of channel occurs where the stream velocity is the highest. Both width and depth increase downstream because discharge increases downstream. As discharge increases the cross sectional shape will change, with the stream becoming deeper and wider. The measurement of the cross section is necessary to determine the total discharge, which is the volume and velocity of the water. Measuring the width and depth of the waterway, and multiplying these measurements together determine the cross section. The depth will vary across the stream and so the width and depth should be measured in small intervals and aggregated to determine the total area.

To determine an average discharge, WV Save Our Streams recommends measuring the depth in at least five positions across the stream, one of these positions being the deepest portion of the channel (thalweg). In most cases the velocity and cross sectional area (width x depth) should be determined from a run. Each time these measurements are made they should be completed from the same section of the reach.

ADDITIONAL SURVEY PROCEDURES

SKETCH THE REACH (the site map): In addition to the observations above, volunteer monitors are asked to sketch a site map of the reach. Draw the site map from direct observation. It should show the main features of the site and their relationship as accurately as possible. As fieldwork continues, modify the map with features such as important habitat conditions (i.e. eroding banks, bars etc.) and indicate the approximate location of water sample and macro-invertebrate sample collection locations. Scale the map to show the entire reach surveyed.

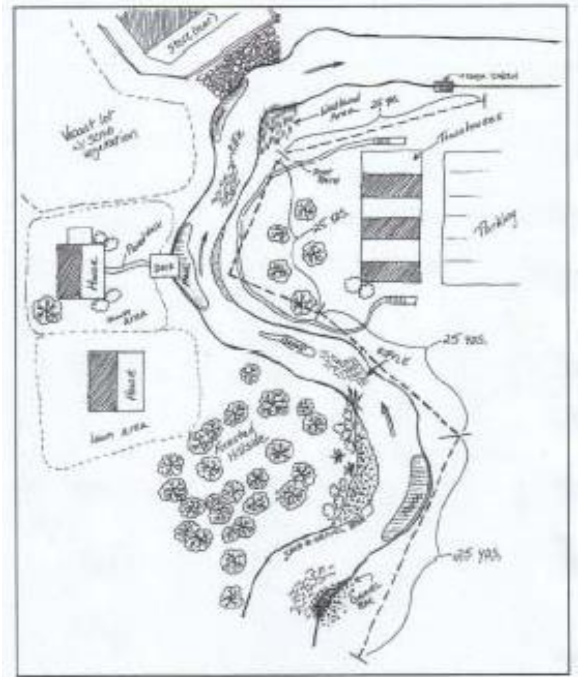


PHOTO DOCUMENTATION: **Photographs** provide a qualitative and potentially semi-quantitative record of conditions in a watershed, or within a stream reach. Photographs can be used to document general conditions, pollution events or other impacts, and document temporal progress for restoration efforts or other projects designed to benefit water quality. Use the same camera to the extent possible for each photo throughout the life of the station. From the inception of any photo documentation until it is completed, always take each photo from the same position (photo point), and at the same bearing and vertical angle. For general reach photo documentation, take at least two photos that show the entire reach. More specific photos can be taken if needed. Try to include landscape features that are unlikely to change over several years (i.e. rocky outcrops, cliffs, large trees, buildings or other permanent structure), so that repeat photos will be easy to position. It is often important to include a ruler, stadia rod or person to convey the scale of the image. Often times an overhead or elevated shot from a bridge, cliff, peak, tree, etc. will be important in conveying the full dimensions.

LAND USE ASSESSMENT: The purpose of this portion of the survey is to get an overall picture of the land surrounding and draining the stream reach. A basic assessment will help you better understand what problems to expect and where to look for those problems. The first step is to review your topographic map and aerial photographs that include your stream stations. Prior to or after completing a stream survey, drive or walk portions of the watershed upstream from your stations to locate any possible activity that may threaten your stream reach. Keep in mind that this rating is simply your judgments of the level of impacts; it is not an actual assessment of the real impacts. The only way to assess a specific impact or activity is to set-up an impact assessment study.

All surveys are mailed to the coordinator so that a proper quality assurance review can occur. After the review is complete the survey data sheets are returned to the volunteer monitoring groups along with a summary of the results, and including other comments or questions the coordinator may have. The data is then entered into the **VOLUNTEER ASSESSMENT DATABASE** (VAD).

REFERENCES

1. Bouchard R.W. 2002. **Guide to Aquatic Invertebrates of the Upper Midwest**. University of Minnesota's Water Resources Center
2. Harrelson, C.C et al. 1994. **Stream Channel Reference Sites: An Illustrated Guide to Field Techniques**. USDA, Forest Service Technical Report RM-245
3. Jessup, B.K., A. Markowitz and J.B. Stribling. 2002. **Family-Level Key to Stream Invertebrates of Maryland and Surrounding Areas**. Tetra Tech, Inc.
4. U.S. EPA, Office of Wetland, Oceans and Watersheds. 1999. **Rapid Bioassessment Protocols for use in Wadeable Streams and Rivers**. EPA 841-B-99-002
5. U.S. EPA, Office of Wetlands, Oceans and Watershed. 1996. **The Volunteer Monitor's Guide to Quality Assurance Project Plans**. EPA 841-B96-003
6. U.S. EPA, Office of Wetlands, Oceans and Watershed. 1997. **Volunteer Monitoring: A Methods Manual**. EPA 841-B97-003
7. Voshell, J.R. 2000. **A Guide to Common Freshwater Invertebrates of North America**. McDonald & Woodward Publishing Company

WV Save Our Streams Program: <http://www.dep.wv.gov/sos>

APPENDIX A - SAFETY PRECAUTIONS

WV Save Our Streams does not require signatures on, or provide any type of liability or **hold-harmless waiver forms** for volunteer monitors. The primary objectives of the program are to provide education; training and resources volunteers need to carry out a water monitoring survey. The safety at the site is the responsibility of the volunteer monitoring group. If the volunteer group fills the need for such forms, they should seek the advice of a trusted legal advisor. However, the program strongly recommends that volunteers develop specific safety preparedness plans and attend safety and outdoor first-aid training on an annual basis. At least one of the persons present during the survey should have up-to-date safety and first-aid training.

1. **Develop a safety plan:** Find out the location and telephone number of the nearest telephone and write it down. Locate the nearest medical center and write down directions on how to get between the center and your site(s) so that you can direct emergency personnel. Have each member of the sampling team complete a medical form that includes emergency contacts, insurance information, and pertinent health information such as allergies, diabetes, epilepsy etc.
2. **Never drink the water in a stream:** Assume it is unsafe to drink, and bring your own water from home. After monitoring, wash your hands with antibacterial soap.
3. **Always monitor with partner(s):** Use a minimum of 2 persons; teams of 3-4 or more people are best; always let someone else know where you are, when you intend to return and what to do if you don't return at the appropriate time.
4. **Have first aid kits handy:** Know any important medical conditions of team members (e.g., heart conditions or allergic reactions to bee stings). It is best if at least one team member has first-aid and CPR certification.
5. **Listen to weather reports:** Never go sampling if severe weather is predicted or if a storm occurs while at the site.
6. **Never wade high water:** Do not monitor if the stream is very swift or at flood stage; adult volunteers should not enter swift-flowing water above waist-deep, unless absolutely necessary, and young volunteers should not enter swift-flowing water just above knee-deep.
7. **Park in a safe location:** If you drive, be sure your car doesn't pose a hazard to other drivers and that you don't block traffic.
8. **Put your wallet and keys in a safe place:** Use a watertight bag you keep in a pouch strapped to your waist. Without proper precautions, wallet and keys might end up downstream.
9. **Never cross private property without the permission of the landowner:** Better yet, sample only at public access points such as bridge or road crossings or public parks. Take along a card identifying you as a volunteer monitor.
10. **Confirm your location:** Prior to visiting your site(s) check maps, and make sure all volunteers are aware using site descriptions and specific directions.
11. **Know what to do if you get bitten or stung:** Watch for irate dogs, wildlife (particularly snakes), and insects such as ticks, hornets, and wasps.
12. **Watch for vegetation in your area that can cause rashes and irritation:** Learn to identify (in all seasons) poison ivy, poison oak, sumac and other plants that may cause irritation; be aware of briars and thorny plants as well.
13. **Do not monitor if the stream is posted as unsafe:** Do not monitor if the water appears to be severely polluted. No matter what the water conditions are; always remove wet shoes and clothes as soon as possible after leaving the stream; use anti-bacterial soap and shower soon after the stream survey is completed.
14. **Do not walk on unstable stream banks:** Disturbing these banks can accelerate erosion and might prove dangerous if a bank collapses. Disturb streamside vegetation as little as possible.
15. **Be very careful when walking in the stream itself:** Rocky-bottom streams can be very slippery and can contain deep pools; muddy-bottom streams might also prove treacherous in areas where mud, silt, or sand has accumulated. If you must cross the stream, use a walking stick to steady yourself and to probe for deep water or muck. Your partner(s) should wait on dry land ready to assist you if you fall. Do not attempt to cross a stream that is swift.
16. **Come prepared for outside work:** Wear clothes such as a hat, loose fitting clothes (especially during warmer weather), closed toed shoes such as sneakers, boots or waders. Felt glued to the bottom of the shoe will improve your traction for walking in the stream. The outside conditions should determine your overall manner of dress. You should also have plenty of drinking water, sunscreen and insect repellent.

A well-equipped **first aid kit** should suffice for most of your medical situations. At a minimum your kit should contain the following items:

1. Telephone numbers of emergency personnel such as the police and ambulance (Know the location of the nearest medical facility and the nearest cell-phone signal.)
2. Several different size Band-Aids for minor cuts.
3. Antibacterial and/or alcohol wipes.
4. First-aid crème or ointments.
5. Triangular bandages and several gauze-pads 3-4 inches square for deeper cuts.
6. Acetaminophen for minor pain relief.
7. A needle and tweezers for removing splinters.
8. Small scissors or a single-edge razor blade for cutting tape to size.
9. A two-inch roll of gauze for large cuts (tunicates).
10. A large compress bandage to hold dressings in place.
11. A three-inch wide elastic bandage for sprains and to aid in applying pressure when necessary to slow bleeding.
12. If participants are allergic to bee stings, include a doctor prescribed antihistamine; make sure all volunteers have their necessary allergy medications prescribed for their specific condition.
13. Always have emergency telephone numbers, contact person(s) and medical information for all participants in case of emergency.

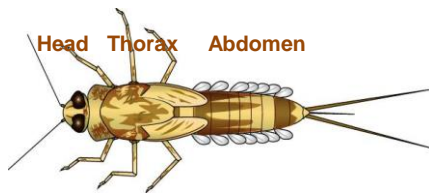
APPENDIX B - EQUIPMENT LIST

In many cases not all of the **equipment** listed is necessary for each monitoring session; however, you should carefully plan your fieldwork and make a checklist of equipment and other items needed prior to your excursions to the stream.

1. Appropriate clothes and foot wear (i.e. hats, loose-fitting clothes for warm weather, layered clothes for cooler weather; raingear for wet-weather; waders, boots, close-toed shoes)
2. Carrying case (i.e. backpacks, plastic containers etc.)
3. Chemical kits and meters (pH meters or kits, dissolved oxygen kit and other chemical monitoring kits as needed)
4. Collection bottles for water samples and vials for macroinvertebrate preservation
5. Collection Nets (usually a two-pole or single-pole kick-net)
6. First aid kit
7. Flow meter or equipment that can be adapted to measure flow such as, an aluminum straight-rod or a float
8. Forceps (tweezers)
9. Gloves (non-allergenic latex or rubber)
10. Open-reel tape measure (100 and 300 feet)
11. Plastic (flexible) mm ruler or gravelometer for pebble counts
12. Preservatives (i.e. alcohol or formalin)
13. Sample size delineator and embeddedness survey ring (optional)
14. Scrub brush (for rock rubbing)
15. Several sizes of buckets: (1) 2 to 3 ½ gallon and at least (1) 5-gallon
16. Sieve such as a wash bucket or 400-micron EZ-strainer, to aid in sorting, counting and separating
17. Small folding table and chairs
18. Sorting trays (white bottom and durable; divided plastic inserts; ice cube trays and plastic craft organizers also work well)
19. Survey instruments (stadia rod etc.) for measuring channel profiles
20. Submersible thermometer
21. Various types of magnifying lenses (box magnifiers, 10^x hand-lens (loupes), etc.)
22. Wash bottles

APPENDIX C – MACROINVERTEBRATE ID GUIDE AND STREAM SURVEY DATA SHEET

WV Save Our Streams Macroinvertebrate ID-Guide

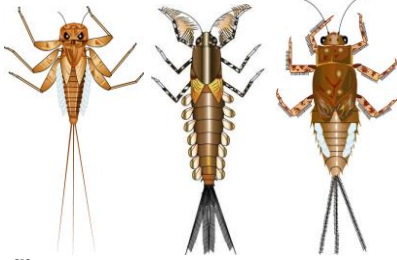


Small minnow mayfly

What is an insect? An insect is an invertebrate (an animal with no spine) that has three-pairs of legs (except Diptera) and three body divisions; the head is the location of the mouth, antenna and eyes; the thorax is the attachment site for the legs and wing pads; and the abdomen, which often has a variety of structures attached including filaments gills and tails. Gills are usually leaf-like, plate-like, or thin filaments. Tails can be long and thin, hairy, webbed or paddle-like. Most of the **benthic macroinvertebrates** you will encounter during stream surveys are aquatic larva or nymphs of insects. Most adult stages are not aquatic but the beetles are the exception. The majority of the insects are described and illustrated on the first page and the top of the second page; non-insect group descriptions and Illustrations begin on the next page.

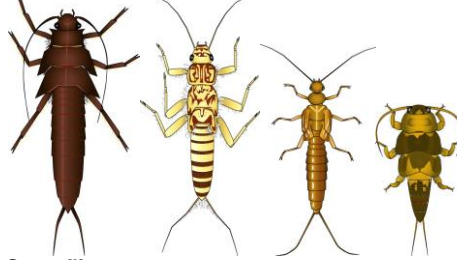
Insect Groups

Instructions provided at the bottom of the next page



Mayflies

(Order **Ephemeroptera**): Three-pairs of legs with a single hook at the end; three some-times two tail filaments; gills attached to the abdomen, which may sometimes be covered and difficult to see. Mayflies exhibit several types of move-ments (or habits); swimmers, clingers, crawlers and burrowers. (VS-M) (M)



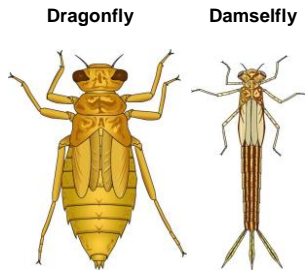
Stoneflies

(Order **Plecoptera**): Three-pairs of legs with two-hooks at the end; two tail filaments; no gills attached to the abdomen but some kinds may have gills near the top of the abdomen; gills if visible, mostly on the legs and thorax. (S-VL) (M)



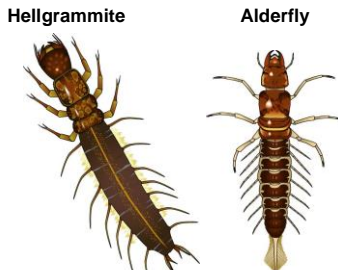
Case-building caddisflies

(Order **Trichoptera**): Grub-like soft body and a hard head; Three-pairs of legs located on the upper third of the body; tail is small and usually forked, sometimes fringed with hairs; gills are scattered on the underside of the abdomen. The case (retreat) is a relatively solid structure made of a variety of stream-bed materials held together by silk. (VS-L) (M)



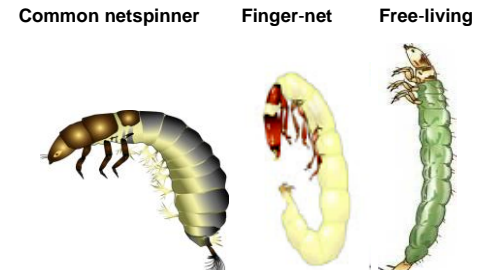
Dragonflies and Damselflies

(Order **Odonata**): Three-pairs of legs; large eyes; long spoon-like jaws; no tails on the abdomen. Dragonflies have a broad shaped abdomen, while the Damselfly abdomen is much narrower. Damselfly gills are attached to the end of the abdomen, they look like tails. (M-VL) (M)



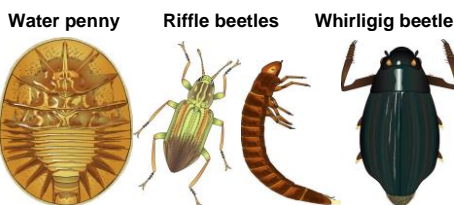
Fishflies and Alderflies

(Order **Megaloptera**): Three-pairs of legs; large pinching jaws; eight-pairs of filaments attached to the sides of the abdomen. Fishflies also called **hellgrammites** have a two-hooked tail, whereas Alderflies have a single tapered tail and are usually much smaller and lighter in color. (M-VL) (M)



Net-spinning caddisflies

(Order **Trichoptera**): Similar characteristics as above but the abdomen usually has more abundant gills, especially the **common netspinner** (family **Hydropsychidae**). The net-spinner's retreat is also made of a variety of streambed materials, which are held together more loosely by fine strands of silk. The **free-living caddisfly** (right) does not build a case or net. (S-L) (M)



Beetles

(Order **Coleoptera**): Three-pairs of legs; body usually covered by a hard exoskeleton. The Most common kinds collected are the **water penny** and **riffle beetles** (left-right), but others kinds are also found. (VS-L) (M)

True flies

(Order **Diptera**): Usually the body is segmented with some type of visible features either along the body, or at the head or tail regions (i.e. head, tails, prolegs, whelps etc.). **This order is the only aquatic insect without fully developed legs in the larval stages.** Dipterans are very diverse order with many aquatic varieties. Several common kinds are described here. (M)



Non-biting midge

(Order **Diptera**; family **Chironomidae**): Segmented body with a visible head; two leg-like projections at the front and rear. Sometimes they are bright **red** in color. (VS-M)



Crane fly

(Order **Diptera**; family **Tipulidae**): No legs, no visible head; plump body with lobes along the segments; may have structures that look like tentacles, lobes or one bulb at the end of the body. (S-VL)



Black fly

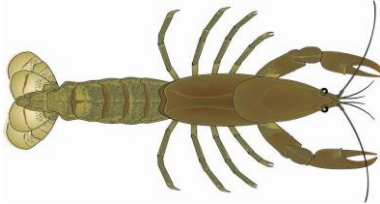
(Order **Diptera**; family **Simuliidae**): Body has a bowling-pen shape (lower is wider than the upper); there are multiple brushes/fans on the head and a ring of hooks on the abdomen. (VS-M)



Watersnipe fly

(Order **Diptera**; family **Athericidae**): Plump body, looks very much like a caterpillar; on the underside there are structures that look similar to legs but are not segmented; the tail is forked and fringed with hairs. (S-L)

Non-Insect Groups



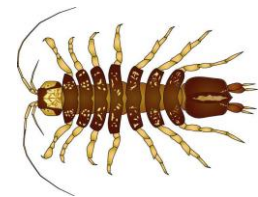
Crayfish

(Class **Crustacea**; order **Decapoda**): Five pairs of legs, the first two usually have large claws; large flipper-like structure at the end of the abdomen. (M-VL)



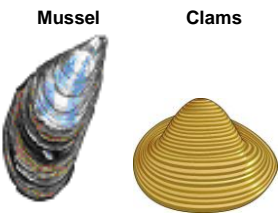
Scud/Sideswimmer

(Class **Crustacea**; order **Amphipoda**): Seven pairs of legs, the first two may be claw-like; body is somewhat higher than it is wide. Usually swims with a sideways motion. (S-M)



Aquatic sowbug

(Class **Crustacea**; order **Isopoda**): Seven pairs of legs, the first two may be claw-like; very long antenna; body is wider than it is high, giving the animal a fairly flattened appearance. (S-M)



Clams and Mussels

(Class **Bivalvia**): Fleshy body enclosed between two-hinged shells; the shape and ridge spacing of the shells can determine different kinds. **Mussels** are usually larger than clams and have dark colored oblong shells. (VS-VL) (M)



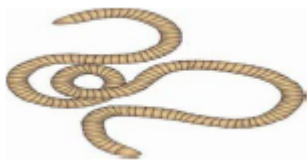
Operculate snails

(Class **Gastropoda**; sub-class **Prosobranchia**): Fleshy body enclosed by a single shell, which is usually coiled in an upward spiral. The opening of the shell is covered by an operculum (door). (VS-L) (M)



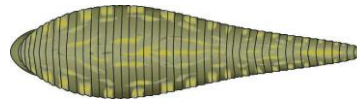
Non-operculate snails

(Class **Gastropoda**; sub-class **Pulmonata**): Fleshy body enclosed by a single shell, which is sometimes coiled upward but also may lie flat or have a conical shape. The opening of the shell is not covered by an operculum. (VS-L) (M)



Aquatic worms

(Phylum **Annelida**; class **Oligochaeta**): Body is long with numerous segments along its entire length; has no visible head or tail. (VS-VL)



Leeches

(Phylum **Annelida**; class **Hirudinea**): Body is long and thin or slightly widened; 34-segments along its length, but there appears to be many more. (S-VL)



Flatworms

(Class **Turbellaria**): Soft elongate body without segment; head triangular shaped with eyes on top, which give the animal a cross-eyed appearance. (VS-L)



Instructions: Identification is easier when the organism is viewed in the same orientation as its illustration. Illustrations are drawn mostly in top and side views; the water penny is shown in underside view. The (M) symbol indicates that multiple kinds may be collected from the group (Order or Class). Use **morphological** features as your basis for identification; the size and color are often variable and influenced by environmental factors. Only a few of the many kinds possible are illustrated. **Sizes illustrated not proportional**

<http://www.dep.wv.gov/sos>

Size categories (mm): > 50 Very large (VL); 50 - 30 Large (L); 29 -10 Medium (M); 10 - 5 Small (S); < 5 Very small (VS)

Illustrations courtesy of the **Cacapon Institute**; Jennifer Gillies, artist



1. DETERMINE THE STREAM-REACH BOUNDARY. 2. NEAR THE LOWER END OF THE REACH (IN THE DEEPEST PORTION OF THE RUN), COLLECT WATER SAMPLES AND ANALYZE USING THE CHEMICAL TESTS YOU HAVE AVAILABLE. YOU MAY USE YOUR COLLECTION CONTAINER TO OBSERVE WATERCOLOR AND CLARITY AND TO DETERMINE WATER ODORS. 3. MEASURE THE WIDTH-DEPTH AND VELOCITY, AND ESTIMATE THE WATER LEVEL. 4. USING A **KICK-NET**, COLLECT A MINIMUM OF THREE BENTHIC MACROINVERTEBRATE SAMPLES FROM THE BEST RIFFLES OR RUNS WITHIN YOUR STREAM REACH. USE THE TALLY SHEET ON PAGE FOUR TO RECORD INFORMATION ABOUT YOUR COLLECTIONS. 5. EVALUATE THE PHYSICAL AND HABITAT CONDITIONS, AND RECORD INFORMATION ABOUT KNOWN LAND USE ACTIVITIES. 6. SKETCH YOUR REACH OR SUBMIT PHOTOGRAPHS WITH THE SURVEY, AND ADD ANY OTHER COMMENTS THAT YOU FEEL ARE IMPORTANT. **NOTE: A SCIENTIFIC COLLECTION PERMIT FROM WVDNR IS REQUIRED FOR ALL BENTHIC SURVEYS.**

Stream name _____ Survey date _____
 Watershed _____ County _____
 Latitude _____ Longitude _____ Directions _____
 Start/end times _____
 Survey completed by _____ Station code _____
 Affiliation _____ E-mail _____
 Mailing address _____ Phone number _____

WATER CHEMISTRY: Use the boxes below to record the results of your water chemistry analysis; attach additional sheets if necessary.

	Result	units		Result	units		Result	units
Temperature (C/F)			Conductivity			Alkalinity		
Dissolved oxygen			Nitrates			Iron		
pH			Turbidity			Fecal/E-coli		
Additional tests (describe and record results) _____								

PHYSICAL CONDITIONS: Use the check boxes below to describe the conditions that closely resemble those of your stream. The extra lines are provided to write in any additional comments. You may see more than one type of condition; if so, be sure to indicate these on your survey (check all that apply). If multiple conditions are observed, always indicate the most dominant condition. Note: If the condition you observe is not listed, describe it in the comment section.

Water clarity	Water color	Water/Sediment odor	Surface foam
		Water	Sediment
Clear	None	None	None
Murky	Brown	Fishy	Slight
Milky	Black	Musky	Moderate
Muddy	Orange/red	Rotten egg	High
Other (describe)	Gray/White	Sewage	
	Green	Chemical	

Algae color	Algae abundance	Algae growth habit	Streambed color
Light green	None	Even coating	Brown
Dark green	Scattered	Hairy	Black
Brown	Moderate	Matted	Green
Other (describe)	Heavy	Floating	White/gray
			Orange/red

Physical condition comments: _____

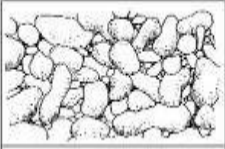
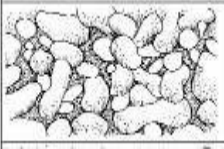


Weather (today and past 48-hours) _____

Indicate the % of your reach that is shaded	> 80 Excellent	80 - 60 Good	60 - 40 Fair	< 40 Poor

WIDTH AND DEPTH MEASUREMENTS: Record the wetted width and average depth from at least two of the channel's habitats (RUN, RIFFLE or POOL). Determine the average depth from a minimum of five measurements (one should always be from the deepest part of the channel). The width should be measured from the widest section of the feature.

1. Riffle	Wetted Width ^(feet) _____	Depth ^(feet) _____
2. Run	Wetted Width ^(feet) _____	Depth ^(feet) _____
3. Pool	Wetted Width ^(feet) _____	Depth ^(feet) _____

HABITAT CONDITIONS: Rate the habitat conditions by choosing the best description for the reach, and then choose a score from the range within the description. Note: Bank stability and riparian buffer width are assessed on both the **LEFT** and **RIGHT** side of the stream.

		20	19	18	17	16	15	14	13	12	11	10	9	8	7	6	5	4	3	2	1
Embeddedness EVALUATED IN RIFFLES																					
		Optimal					Suboptimal					Marginal					Poor				
Sediment deposition		Little or no formation of depositional features; < 20% of the reach affected. See below for examples					Some increase in depositional features; 20-40% of the reach affected.					Moderate amounts of depositional features; 40-60% of the reach affected.					Heavy amounts of deposition; > 60% of the reach affected.				
		Optimal					Suboptimal					Marginal					Poor				

The next two conditions are evaluated on both the left and the right sides of the stream. Note: The **LEFT** and **RIGHT** sides are determined by looking downstream.

Bank stability				10	9	8	7	6	5	4	3	2	1
				Banks are stable; no evidence of erosion or bank failure; little or no potential for future problems; < 10% of the reach affected.		Banks are moderately stable; infrequent areas of erosion occur, mostly shown by banks healed over or a few bare spots; 10-30 % of the reach affected.		Banks are moderately unstable; 30-50% of the reach has some areas of erosion; high potential for erosion during flooding events.		Banks are unstable; many have eroded areas (bare soils) along straight sections or bends; obvious bank collapse or failure; > 50% affected.			
Left		Right		Optimal		Suboptimal		Marginal		Poor			
Riparian buffer width				Mainly undisturbed vegetation > 60 ft; no evidence of human impacts such as parking lots, road beds, clear-cuts, mowed areas, crops, lawns etc.		Zone of undisturbed vegetation 40-60 ft; some areas of disturbance evident.		Zone of undisturbed vegetation 20-40 ft; disturbed areas common throughout the reach.		Zone of undisturbed vegetation < 20 ft; disturbed areas common throughout the entire reach.			
				Optimal		Suboptimal		Marginal		Poor			
Left		Right		Optimal		Suboptimal		Marginal		Poor			

Totals		> 70	70 - 55	54 - 40	< 40
		Optimal	Suboptimal	Marginal	Poor

Habitat condition comments: _____

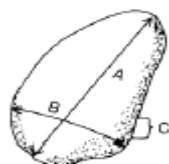
SEDIMENT DEPOSITION may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends.

NOTE: TO CALCULATE A SCORE BASED ON A 0-100 SCALE DIVIDE THE TOTAL BY 80 AND MULTIPLY BY 100

STREAMBED COMPOSITION: You should always collect information about the composition of your reach. You can either estimate the proportions or you use a **PEBBLE COUNT** for a more accurate measure of composition. At a minimum you should estimate composition of the riffles within your reach. The size categories are determined by the (B) axis measured in millimeters. Use the table below to record your data. Did you estimate ☐ or count? ☐

Silt/clay < 0.06	Sand 0.06 – 2	Gravel 2 - 24 25 - 64	Cobble 65 - 255	Boulder 256 - 1096	Bedrock > 1096	Woody debris
Very small; having a smooth slick feel	Very small; having a grainy feel	Pea to tennis ball Fine Coarse	Tennis ball to basketball	Basketball to car size	Usually larger than a car; solid surface	Includes sticks, leaves etc

Riffle only ☐ Entire reach ☐ Estimates should be made from riffles only



- (A) Long axis (**Length**)
(B) Intermediate axis (**Width**)
(C) Short axis (**Height**)

Pebble counts require two people, one in the stream and one on shore. The person in the stream walks upstream from bank to bank using a zigzag pattern. After each step the person reaches down without looking, picks up the first particle touched, and measures the intermediate axis with a ruler. The on-shore partner records the measurement. The process continues until **100** pebbles have been measured or the reach has been walked. For a quick estimate, the coordinator recommends that **50** be collected from the entire reach and **20** if collecting from riffles only. You should divide the gravel category into fine and coarse to get a more accurate measure. Note: Pebble counts are not required; they are optional and should only be completed once each year or less frequently.

LAND USE: Indicate the land uses that you believe may be having an impact on your stream station. Use the letters (**S**) streamside, (**M**) within ¼ mile and (**W**) somewhere in the watershed, to indicate the approximate location of the disturbance and the numbers (**1**) slight, (**2**) moderate or (**3**) high, to represent the level of disturbance.

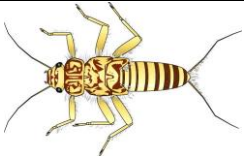
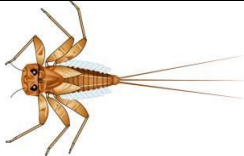

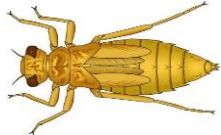
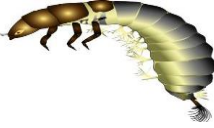

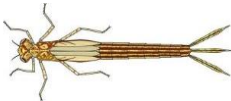




















Active construction			Pastureland			Single-family residences		
Mountaintop mining			Cropland			Sub-urban developments		
Deep mining			Intensive feedlots			Parking lots, strip-malls etc.		
Abandoned mining			Unpaved Roads			Paved Roads		
Logging			Trash dumps			Bridges		
Oil and gas wells			Landfills			Other (describe)		
Recreation (parks, trails etc.)			Industrial areas					

Pipes? ☐ Yes ☐ No

Describe the types of pipes observed and indicate if there is any discharge from the pipes. Also describe the colors and odors of the discharge, and provide any other land-use comments _____

PHOTOGRAPH AND SKETCH THE STUDY REACH: Use the space below to draw a **bird's eye view** your study reach. Indicate the direction of flow, sample locations and important features of the reach. Choose at least two locations from which to take your photos and submit your photos with your survey data sheet.

BENTHIC MACROINVERTEBRATES: Use the table below to record information about your collections. Record their abundance using these codes: **(A)** > 50, **(C)** 5 – 50 and **(R)** < 5 and also record the number of different kinds. The # of kind's box indicates groups in which multiple kinds (**FAMILIES**) are possible. Note: Always record the **# OF KINDS** when necessary. Illustrations courtesy of the [Cacapon Institute](#); Jennifer Gillies, artist

 Stoneflies	<input type="text"/>	 Mayflies	<input type="text"/>	 Caddisflies	<input type="text"/> Case-builders
 Dragonflies	<input type="text"/>	 Common netspinner	<input type="text"/>	 Caddisflies	<input type="text"/> Net-spinners <input type="text"/> Free-living
 Damselflies	<input type="text"/>	 Riffle beetle	<input type="text"/>	 Water penny	<input type="text"/>
 Fishfly/Hellgrammite	<input type="text"/>	 Alderfly	<input type="text"/>	 Other Beetles/Bugs	<input type="text"/> Beetles <input type="text"/> True bugs
 Midges	<input type="text"/>	 Black fly	<input type="text"/>	 Crane fly	<input type="text"/>
 Watersnipe fly	<input type="text"/>	 Other True flies	<input type="text"/>	 Crayfish	<input type="text"/>
 Clams	<input type="text"/>	 Mussel	<input type="text"/>	 Scud/Sideswimmer	<input type="text"/>
 Operculate snails	<input type="text"/>	 Non-operculate snails	<input type="text"/>	 Aquatic sowbug	<input type="text"/>
 Aquatic worm	<input type="text"/>	 Leech	<input type="text"/>	 Flatworm	<input type="text"/>

Other aquatic life observed or collected: _____

STREAM SCORE

After the sorting and identifications is complete, the macroinvertebrates are assessed using four **metrics**. First, transform your abundance rating into numbers using this code (**A = 6; C = 3; R = 1**) and follow the instructions below to complete all calculations. **Note:** The **SHADING** indicates that multiple kinds are possible within the group.

1. **Biotic Index:** Multiply the abundance number by the tolerance value to calculate the tolerance score. Add the entire tolerance score column and the abundance column. Divide the tolerance total by the abundance total.
2. **Total Taxa:** Calculate the total number of kinds.
3. **EPT Taxa:** Calculate the total number of kinds from the stoneflies, mayflies, and all caddisflies.

The final step is to determine a **point value** for each metric. These points are added together to determine your overall **stream score** and integrity rating. **Note: Don't forget to record the number of kinds.**

BENTHIC MACROINVERTEBRATES	Abundance	Tolerance Value	Tolerance Score	Number of Kinds
Stoneflies (Order Plecoptera)		2		
Mayflies (Order Ephemeroptera)		3		
Case-building caddisflies (Order Trichoptera)		3		
Net-spinning caddisflies (Order Trichoptera)		4		
Common netspinner (Family Hydropsychidae)		5		
Free-living caddisfly (Family Rhyacophilidae)		3		
Dragonflies (Sub-order Anisoptera)		4		
Damselflies (Sub-order Zygoptera)		7		
Riffle beetle (Family Elmidae)		4		
Water penny (Family Psephenidae)		3		
Other Beetles (Order Coleoptera)		6		
True Bugs (Order Hemiptera)		8		
Hellgrammite (Family Corydalidae)		3		
Alderfly (Family Sialidae)		6		
Non-biting midge (Family Chironomidae)		9		
Black fly (Family Simuliidae)		6		
Crane fly (Family Tipulidae)		4		
Watersnipe fly (Family Athericidae)		3		
Other True flies (Order Diptera)		7		
Water mite (Order Hydrachnida)		6		
Crayfish (Family Cambaridae)		5		
Sideswimmer (Family Gammaridae)		5		
Aquatic sowbug (Family Asellidae)		7		
Operculate snails (Sub-class Prosobranchia)		5		
Non-operculate snails (Sub-class Pulmonata)		7		
Clams (Order Veneroida)		6		
Mussel (Family Unionidae)		4		
Aquatic worm (Class Oligochaeta)		10		
Leech (Class Hirudinea)		10		
Flatworm (Class Turbellaria)		7		
Other invertebrates (describe)	Total Abundance		Total Tolerance	Total Taxa (# OF KINDS)

Metrics	Results	Points	10	8	6	4	2
1. Total Taxa			> 18	18 - 15	14 - 11	10 - 7	< 7
2. EPT Taxa			> 10	10 - 8	7 - 5	4 - 2	< 2
3. Biotic Index			< 3.5	3.5 - 4.3	4.4 - 5.6	5.7 - 6.5	> 6.5

Integrity Rating Scale

STREAM SCORE

> 24	24 - 19	18 - 13	< 13
Optimal	Suboptimal	Marginal	Poor

Tolerance values (TVs): TVs are general numbers that represent the tolerance of most of the group. It should be noted that tolerance ranges vary for certain Mayflies (a few kinds range from 4-5), Beetles (a few kinds are 5), other True flies (a few kinds are 6; Net-wing midge TV = 2) and Operculate snails (a few kinds are less than 5).

DISCHARGE

Determine the discharge by using a flow meter or other methods such as the **Float** or the **VELOCITY HEAD ROD (VHR)** method. The more measurements collected the more accurate your discharge results will be; however, you should collect a minimum of five measurements. Discharge should always be measured from a **RUN**. Stretch your tape measure across the run and select a minimum of five positions along the tape to measure discharge. One measurement should be from the deepest part of the channel and the others should be on either side. If you use the float method move 10-20 feet upstream from the tape and float at least five times back to the tape. The float distance must be timed in seconds.

Discharge method used

Water Level

☐

Float

☐

VHR

☐

Flow meter

☐

Low

☐

Normal

☐

High

☐

Dry

Channel width _____ feet

Tape positions (ft)	Depth (ft)	Velocity (ft/sec)	VHR (Rise-inches)	Float (sec)	Discharge (cfs)
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
Totals/Averages					

Cross Sectional Area (CSA) _____ ft²

(CSA = Average Depth x Width)

Discharge = CSA x Velocity

= _____ x _____
= _____ cfs (ft³/sec)

If you use a float record your distance below and the number of seconds it took to travel the distance in the column indicated. **Float distance** (feet) _____

VHR rises and velocities

Rise (R)	Velocity	Rise (R)	Velocity
¼	1.2	3 ¼	4.2
½	1.6	3 ½	4.3
¾	2.0	3 ¾	4.5
1	2.3	4	4.6
1 ¼	2.6	4 ¼	4.8
1 ½	2.8	4 ½	4.9
1 ¾	3.1	4 ¾	5.0
2	3.3	5	5.2
2 ¼	3.5	5 ¼	5.3
2 ½	3.7	5 ½	5.4
2 ¾	3.8	5 ¾	5.5
3	4.0	6	5.7

VHR Velocity = $8 \times \sqrt{R}$, where R is rise in feet

Submit a clear copy or the original data sheet to the coordinator at address below. The Coordinator will review your survey and return it with comments or return a summary with comments. **ALWAYS KEEP A COPY FOR YOUR RECORDS.**

West Virginia Dept. of Environmental Protection
Save Our Streams Program
601 57th Street, SE
Charleston, WV 25304

For more information visit the program's website at: <http://www.dep.wv.gov/sos>. A summary data sheet is provided on the next page.

Stream name _____ Basin _____
 Monitoring group _____ Station name _____
 County _____ Latitude

--	--	--

 Longitude

--	--	--

 Survey date _____ Directions _____
 Database code _____ RR miles _____

WATER CHEMISTRY

	Units
Temperature (C/F)	
pH	
Conductivity	µ/cm
Dissolved Oxygen	
Nitrate/Nitrite	
Alkalinity	
Acidity	
Turbidity	
Bacteria (Fecal/E-coli)	CFU

Other (Describe and record the results below)

PHYSICAL CONDITIONS

Run width ^{ft}		Run depth ^{ft}	
Riffle width ^{ft}		Riffle depth ^{ft}	
Pool width ^{ft}		Pool depth ^{ft}	

Water level	
Discharge (cfs)	
Water clarity	
Water color	
Water/Sediment odor	
Sediment color	
Surface foam	
Algae color	
Algae abundance	
Algae texture	
Channel shade	

COMPOSITION

	Estimate	Count
Silt/clay		
Sand		
Gravel (fine/coarse)		
Cobble		
Boulder		
Bedrock		
Woody debris		
Index		

Evaluate the streambed composition using a **PEBBLE COUNT** procedure whenever possible, or simply estimate the percentages.

HABITAT CONDITIONS

Sediment deposition	
Embeddedness	
	Left Right
Bank stability	
Riparian buffer width	
Total score	
Integrity Rating	

Note: Habitat conditions are evaluated on a scale of 1-20 and 1-10

BENTHIC MACROINVERTEBRATES

	Abundance	Taxa
Mayflies (<i>Ephemeroptera</i>)		
Stoneflies (<i>Plecoptera</i>)		
Case-building caddisflies (<i>Trichoptera</i>)		
Net-spinning caddisflies (<i>Trichoptera</i>)		
Free-living caddisfly (<i>Rhyacophilidae</i>)		
Common netspinner (<i>Hydropsychidae</i>)		
Dragonflies (<i>Anisoptera</i>)		
Damselflies (<i>Zygoptera</i>)		
Riffle beetle (<i>Elmidae</i>)		
Water penny (<i>Psephenidae</i>)		
Beetles/Bugs (<i>Coleoptera/Hemiptera</i>)		
Hellgrammite/Fishfly (<i>Corydalidae</i>)		
Alderfly (<i>Sialidae</i>)		
Non-biting midge (<i>Chironomidae</i>)		
Black fly (<i>Simuliidae</i>)		
Crane fly (<i>Tipulidae</i>)		
Watersnipe fly (<i>Athericidae</i>)		
Other True flies (<i>Diptera</i>)		
Water mite (<i>Hydrachnidae</i>)		
Crayfish (<i>Cambaridae</i>)		
Scud/Sideswimmer (<i>Gammaridae</i>)		
Aquatic sowbug (<i>Asellidae</i>)		
Clams (<i>Veneroida</i>)		
Mussel (<i>Unionidae</i>)		
Operculate snails (<i>Prosobranchia</i>)		
Non-operculate snails (<i>Pulmonata</i>)		
Aquatic worms (<i>Oligochaeta</i>)		
Leeches (<i>Hirudinea</i>)		
Flatworms (<i>Turbellaria</i>)		
Other		

> 50 (Abundant) = 6; 5-50 (Common) = 3; < 5 (Rare) = 1

TOTALS

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List other aquatic animals collected or observed

Habitat and physical condition comments

BIOLOGICAL INTEGRITY

	Value	Points
1. Total Taxa		
2. EPT Taxa		
3. Biotic Index		
Stream score		
Integrity Rating		

LAND USE CONDITIONS

Additional comments: _____



WEST VIRGINIA DEPARTMENT OF ENVIRONMENTAL PROTECTION

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